Composition and physical properties of cress (*Lepidium sativum* L.) and field pennycress (*Thlaspi arvense* L.) oils[☆]

Bryan R. Moser[*], Shailesh N. Shah, Jill K. Winkler-Moser, Steven F. Vaughn, Roque L. Evangelista

United States Department of Agriculture, Agricultural Research Service, National Center for Agricultural Utilization Research, 1815 N. University St., Peoria, IL 61604, USA

ABSTRACT

The fatty acid profiles and tocopherol and phytosterol contents of crude oils of cress (*Lepidium sativum* L.) and field pennycress (*Thlaspi arvense* L.) are reported, along with yields from the corresponding seeds. The physical properties of these oils were also determined, which included oxidative stability, kinematic viscosity, viscosity index, low temperature fluidity, specific gravity, acid value, lubricity, and iodine value. The oil content of dried cress and field pennycress seeds was 22.7 and 29.0 wt%, respectively. The primary fatty acids found in cress oil were oleic (30.6 wt%) and linolenic acids (29.3 wt%), whereas field pennycress oil was principally composed of erucic (32.8 wt%) and linoleic (22.4 wt%) acids. Cress oil contained high concentrations of γ- (1422 ppm) and δ- (356 ppm) tocopherols, whereas α-tocopherol (714 ppm) was the primary tocopherol discovered in field pennycress oil. The overall tocopherol concentrations of cress and field pennycress oils were 1799 and 851 ppm, respectively. The primary phytosterols elucidated in cress and field pennycress oils were sitosterol and campesterol, with avenasterol also present in significant quantity in cress oil. The total phytosterol concentration in cress oil (14.41 mg/g) was greater than that in field pennycress (8.55 mg/g) oil. Field pennycress oil exhibited excellent low temperature fluidity, whereas cress oil was more stable to oxidation and over a range of temperatures displayed lower kinematic viscosities as well as a higher viscosity index. The acid and iodine values of field pennycress oil were lower than those for cress oil, but both oils had excellent lubrication properties.

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1. Introduction

Cress (*Lepidium sativum* L.), otherwise known as garden cress, gardencress pepperweed or garden pepperwort, is a fast growing annual herb belonging to the Brassicaceae family that is native to Egypt and west Asia but is widely cultivated in temperate climates throughout the world for various culinary and medicinal uses (Gokavi et al., 2004). Field pennycress (*Thlaspi arvense* L.), also known as stinkweed, is a winter annual also belonging to the Brassicaceae family that is native to Europe but has a wide distribution throughout temperate North America (Vaughn et al., 2005). Generally considered to be an agricultural pest (weed), field pennycress can serve in a summer/winter rotational cycle with conventional commodity crops (such as soybean), thus not displacing existing agricultural production. Advantages to cress and field pennycress include relatively high oil contents, tolerance of fallow lands, min-

imal agricultural inputs (pesticides, fertilizer, water), capability to serve in rotational crop cycles, and compatibility with existing farm infrastructure. The reported yield of oil from seeds of cress and field pennycress is 23 and 29 weight percent (wt%), respectively (Dolya et al., 1976).

The objective of the current study was to evaluate the chemical compositions and physical properties of cress and field pennycress oils and to compare those results with soybean oil. Specifically, the fatty acid (FA) compositions and phytosterol and tocopherol contents were determined, along with oxidative stability, low temperature fluidity, kinematic viscosity, viscosity index, lubricity, iodine value, specific gravity, and acid value.

2. Materials and methods

2.1. Materials

Cress seeds (cv. 'Cressida') were purchased from Johnny's Selected Seeds (Winslow, ME). Field pennycress seeds were collected from a wild population in Peoria County, IL. Refined, bleached, and deodorized (RBD) soybean oil which did not contain commercial additives was purchased from KIC Chemicals, Inc. (New Platz, NY) and was used as received. Stigmasterol and 5α-

cholestane were purchased from Matreya, Inc. (Pleasant Gap, PA). Campesterol and brassicasterol were purchased from Steraloids (Newport, RI). Each phytosterol standard was ≥97% purity. N,O-Bis(trimethylsilyl)-fluoroacetamide with 1% trimethylchlorosilane (BSTFA + 1% TMCS) was purchased from Regis (Morton Grove, IL). All other chemicals and reagents were obtained from Aldrich Chemical Company (Milwaukee, WI) and used as received.

2.2. Cress oil extraction

Cress seeds were ground in a coffee grinder and oil was extracted with hexane for 24 h in a Soxhlet apparatus. Hexane was removed from the oil by rotary evaporation under reduced pressure. For determination of total oil content, 10.0 g triplicates of ground seeds were extracted for 24 h with a Soxhlet apparatus, and after hexane was removed under reduced pressure the weights of the residual oils were calculated.

2.3. Field pennycress oil extraction

Field pennycress seeds were cold pressed using a heavy duty laboratory screw press (Model L250, French Oil Mill Machinery Company, Piqua, OH). The details of this expeller are available elsewhere (Evangelista, 2009). The cored main barrel (or cage) had a diameter of 8.9 cm and a length/diameter (L/D) ratio of 8.3. The compression ratio of the screw configuration employed was 4. Once extracted, the crude oil was filtered to remove solid material. Quantification of total oil content was accomplished largely as described in Section 2.2 utilizing triplicate Soxhlet extractions with hexane.

2.4. Equipment and methods

2.4.1. Fatty acid profile by GC

Fatty acid methyl esters (FAME) were separated using a Varian (Walnut Creek, CA) 8400 GC equipped with an FID detector and SP2380 (Supelco, Bellefonte, PA) column (30 m × 0.25 mm i.d., 0.20 µm film thickness). Carrier gas was He at 1 mL/min. The oven temperature was initially held at 150 °C for 15 min, then increased to 210 °C at 2 °C/min, followed by an increase to 220 °C at 50 °C/min, which was then held for 10 min. The injector and detector temperatures were set at 240 and 270 °C, respectively. FAME peaks were identified by comparison to the retention times of known reference standards. Each FAME determination was run in triplicate and average values are reported.

2.4.2. Tocopherol content by HPLC

Tocopherols were quantified by HPLC according to AOCS official method Ce 8-89 (AOCS, 1999a). Samples were diluted in hexane to a concentration of 50–100 mg/mL, filtered through 0.45 µm centrifugal filters and analyzed by a Varian HPLC Pro-Star model 230 pump, model 410 autosampler, and model 363 fluorescence detector using excitation and emission wavelengths of 290 and 330 nm, respectively. The mobile phase consisted of hexane:2-propanol (99.5:0.5, v/v, made fresh daily) pumped at a rate of 1 mL/min. Samples were injected by autosampler using the full loop option (100 µL), and tocopherols were separated using an Inertsil (Varian) silica column (5 µm, 150 Å, 250 mm × 4.6 mm i.d.). Tocopherol peaks were identified by comparison to the retention times of known reference standards. A mixture of α-, β-, γ-, and δ-tocopherol standards was injected on each day of analysis to verify HPLC response. Samples were quantified using external standard curves. Each determination was run in triplicate and mean values are reported.

2.4.3. Phytosterol content by GC

Oil samples were saponified and phytosterols were extracted essentially as described by (Dutta and Normén, 1998). Approxi-

mately 50 mg of oil with added 5α-cholestane (100 µg, internal standard) was saponified in 2N ethanolic KOH. Non-saponifiable material was extracted twice with hexane and the solvent was removed from the combined hexane fractions under a gentle stream of N_2. Trimethylsilyl (TMS) derivatives of the phytosterols were made by adding 100 µL each of pyridine and BSTFA + 1%TMCS, and heating at 60 °C for 1 h on a heating block. After derivatization, phytosterols were manually injected onto a Varian 3400 GC equipped with an FID and a Supelco SPB^{TM}-1701 (30 m × 0.25 mm × 0.25 µm) capillary column. Helium was used as the carrier gas with a 1:50 injector split. The injector and detector temperatures were 270 and 290 °C, respectively. The column oven initial temperature was 250 °C for 0.5 min, increased at 10 °C/min to 270 °C and held for 27 min, then increased at 10 °C/min to 280 °C and held for 3.5 min. Data collection and integration were performed using Varian Star Chromatography Software Ver. 5.3. Phytosterols were identified by comparison of their retention times (relative to the internal standard, 5α-cholestane) with those of commercially available standards. Phytosterols without commercially available standards, such as δ5-avenasterol, were identified by their relative retention times compared to literature (Dutta and Normén, 1998), and by comparison with samples known to contain those phytosterols. Quantification was carried out by the internal standard method developed with available standards. For phytosterols with no available commercial standard, the response factor for β-sitosterol was used for quantification.

2.4.4. Low temperature operability

Cloud point (CP, °C) and pour point (PP, °C) determinations were made in agreement with ASTM D5773 (ASTM, 2007) and ASTM D5949 (ASTM, 2001), respectively, using a model PSA-70S Phase Technology Analyzer (Richmond, B.C., Canada). Cloud and pour points were rounded to the nearest whole degree (°C). For a greater degree of accuracy, PP measurements were done with a resolution of 1 °C instead of the specified 3 °C increment. Each experiment was run in triplicate and mean values are reported.

2.4.5. Kinematic viscosity, viscosity index, and specific gravity

Kinematic viscosity (v, mm^2/s) was determined with Cannon-Fenske viscometers (Cannon Instrument Co., State College, PA) at 25, 40 and 100 °C in accordance to ASTM D445 (ASTM, 2006). The viscosity index (VI) was calculated from kinematic viscosity data (40 and 100 °C) according to ASTM D2270 (ASTM, 2004a). Specific gravity (SG) was determined at 25 and 40 °C by AOCS official method Cc 10a-25 (AOCS, 1999b) using a Kimax 25 mL gravity pycnometer from Kimble Chase Life Science and Research Products (Vineland, NJ). All experiments were run in triplicate and mean values are reported.

2.4.6. Lubricity

Lubricity (lub, µm) determinations were performed at 60 °C (±1 °C) according to ASTM standard D6079 (ASTM, 2004b) using a high-frequency reciprocating rig (HFRR) lubricity tester (PCS Instruments, London, England) via Lazar Scientific (Granger, IN). Reported wear scar (µm) values were the result of measuring the maximum lengths of the x- and y-axes of each wear scar with a Prior Scientific (Rockland, Massachusetts, USA) Epimat model M4000 microscope, followed by calculating the average of these maximum values. All experiments were run in duplicate and mean values are reported.

2.4.7. Oxidative stability

Induction period (IP, h) was measured following the European Committee for Standardization (CEN) official method EN 14112 (CEN, 2003) at 110 °C utilizing a Metrohm USA, Inc. (Riverview, FL) model 743 Rancimat instrument. The flow rate of air through 3 ± 0.01 g of sample was 10 L/h. The block temperature was 110 °C

with a correction factor (ΔT) of 1.5 °C. The conductivity measuring vessel contained 50 ± 0.1 mL of deionized water. Each sample was run in triplicate and mean values are reported. IP was mathematically determined as the inflection point of a computer-generated plot of conductivity (μS/cm) of deionized water versus time (h).

2.4.8. Acid and iodine values

Acid value (AV, mg KOH/g) titrations were performed as described in AOCS official method Cd 3d-63 (AOCS, 1999c) using a Metrohm 836 Titrando autotitrator equipped with a model 801 stirrer, a Metrohm Solvotrode electrode, and Tiamo 1.1 Light software. However, the official method was modified for scale to use 2 g of sample and 0.02 M KOH. The titration endpoint was determined by the instrument and visually verified using a phenolphthalein indicator. Each sample was run in triplicate and mean values are reported. Iodine value (IV, g I_2/100 g) was calculated from the fatty acid profile according to AOCS official method 1c-85 (AOCS, 1999d).

2.4.9. Average molecular weight of vegetable oils

The average calculated molecular weight (MW$_{calc}$, g/mol) was determined by a weighted average method utilizing the FA profiles depicted in Table 1. Specifically, the molecular weight of each FA found in the vegetable oil was multiplied by its corresponding weight percentage as determined by GC (Section 2.4.1). The sum of these values (minus the acidic proton) was multiplied by three and the glycerol fragment (minus the oxygen atoms, as they were accounted for in the FA fragments) was added, resulting in an average calculated MW of vegetable oil. For the sake of providing calculated MW values that were not artificially low, unknown constituents were assumed to be oleic acid in the case of cress oil and erucic acid in the case of field pennycress oil.

3. Results and discussion

3.1. Fatty acid composition and free fatty acid content

The primary FA found in cress oil were oleic (C18:1; 30.6 wt%) and linolenic (C18:3; 29.3 wt%) acids, with gondoic (C20:1; 11.1 wt%), palmitic (C16:0; 9.4 wt%) and linoleic (C18:2; 7.6 wt%) acids also detected in significant quantities (Table 1). Erucic (C22:1; 3.0 wt%), stearic (C18:0; 2.8 wt%), and arachidic (C20:0; 2.3 wt%) acids were among the minor FA found in cress oil. These results are in close agreement with previous studies (Dolya et al., 1976; Gokavi et al., 2004). The average MW of cress oil calculated from the FA profile was 891.94 g/mol (Table 2). The oil content of dried cress seeds was 22.7 wt% (Table 2), which was similar to that reported (23.08 wt%) in a previous study (Dolya et al., 1976) and close to

Table 1
Fatty acid compositions (wt%) of cress, field pennycress, and soybean oils.

Purity:oil	Crude cress	Crude pennycress	RBD soybean
C14:0[a]	0.1	0.1	0.1
C16:0	9.4	3.1	11.4
C16:1 Δ9	0.3	0.2	0.1
C18:0	2.8	0.5	3.9
C18:1 Δ9	30.6	11.1	22.1
C18:1 Δ11	1.4	1.5	1.6
C18:2 Δ9, 12	7.6	22.4	52.4
C18:3 Δ9, 12, 15	29.3	11.8	7.4
C20:0	2.3	0.3	0.2
C20:1 Δ11	11.1	8.6	0.2
C20:2 Δ11, 14	0.3	1.6	
C20:3 Δ11, 14, 17	0.5		
C22:0	0.6	0.6	0.3
C22:1 Δ13	3.0	32.8	
C22:2 Δ13, 16		0.7	
C22:3 Δ13, 16, 19		0.3	
C24:1 Δ15		2.9	
Unknown (sum)	0.7	1.5	0.3
Σsat[b]	15.2	4.6	15.9
Σmonounsat[c]	46.4	55.6	24.0
Σpolyunsat[d]	37.7	38.3	59.8

[a] The first number indicates the length of the fatty acid chain and the second the number of double bonds (all *cis*) with Δ signifying the location of the double bond(s).
[b] Σsat = 14:0 + 16:0 + 18:0 + 20:0 + 22:0.
[c] Σmonounsat = 16:1 + 18:1 + 20:1 + 22:1 + 24:1.
[d] Σpolyunsat = 18:2 + 18:3 + 20:2 + 20:3 + 22:2 + 22:3.

the range typically reported for soybeans (18–22 wt%; Dornbos and Mullen, 1992; Chung et al., 2003).

Erucic (32.8 wt%), linoleic (22.4 wt%), and linolenic (11.8 wt%) acids were the primary FA detected in field pennycress oil (Table 1). Other FA of significance included oleic (11.1 wt%), gondoic (8.6 wt%), and nervonic (C24:1; 2.9 wt%) acids. The overall level of saturated FA found in field pennycress oil was low (4.6 wt% total), with palmitic acid (3.1 wt%) comprising the majority of the saturated constituents. These results concur with those previously reported (Dolya et al., 1976). The average MW of field pennycress oil calculated from the FA profile was 956.39 g/mol (Table 2), which was higher than that obtained for cress and soybean (873.33 g/mol) oils largely as a result of its greater erucic acid content. Dried field pennycress seeds contained 29.0 wt% oil (Table 2), which was similar to a previous study that indicated the oil content of field pennycress seeds was 29.18 wt% (Dolya et al., 1976).

As seen upon comparison of the FA compositions of cress and field pennycress oils, cress oil contained considerably more saturated FA than field pennycress oil (15.2 wt% versus 4.6 wt%, Table 1). Field pennycress had a greater abundance of monounsaturated

Table 2
Yields and physical properties of cress, field pennycress, and soybean oils.

Purity:oil	Crude cress	Crude pennycress	RBD soybean
Oil content (wt%)	22.7 (0.4)[a]	29.0 (0.4)	18–22[b]
MW$_{calc}$ (g/mol)	891.94	956.39	873.33
CP (°C)	3 (1)	−25 (1)	−7 (1)
PP (°C)	−11 (1)	−28 (1)	−9 (1)
IP, 110 °C (h)	12.3 (1.7)	5.0 (0.1)	8.3 (0.2)
υ, 25 °C (mm^2/s)	44.12 (0.02)	70.01 (0.04)	52.11 (0.09)
υ, 40 °C (mm^2/s)	28.35 (0.05)	40.97 (0.03)	31.49 (0.03)
υ, 100 °C (mm^2/s)	7.35 (0.06)	9.39 (0.01)	7.67 (0.01)
VI	244	224	228
SG, 25 °C	0.912 (0.001)	0.913 (0.001)	0.919 (0.001)
SG, 40 °C	0.902 (0.002)	0.904 (0.001)	0.910 (0.001)
Lub, 60 °C (μm)	133 (4)	125 (5)	124 (2)
AV (mg KOH/g)	2.60 (0.03)	0.61 (0.03)	0.03 (0.03)
IV	130	118	130

[a] Values in parentheses are the standard deviation from the reported mean.
[b] From Dornbos and Mullen (1992) and Chung et al. (2003).

FA than cress oil (55.6 wt% versus 46.4 wt%), but the polyunsaturated content was nearly equivalent (38.3 wt% versus 37.7 wt%). As is common among members of the Brassicaceae family, erucic acid was particularly abundant in field pennycress oil (32.8 wt%), whereas cress oil only contained 3.0 wt% of this constituent. These oils had divergent FA profiles from soybean oil, which contained a very small percentage of FA with 20 carbons or greater (0.7 wt% total, Table 1). The primary FA constituents found in soybean oil included linoleic (52.4 wt%), oleic (22.1 wt%), palmitic (11.4 wt%), and linolenic (7.4 wt%) acids, which was in close agreement with previous reports (Moser, 2008a).

The percentage of free fatty acids (FFA) in crude field pennycress oil was relatively small, as evidenced by an AV of 0.61 mg KOH/g (FFA content = 0.3 wt%) (Table 2). Crude cress oil contained a higher percentage of FFA (1.3 wt%), as indicated by an AV of 2.60 mg KOH/g. Refined, bleached, and deodorized (RBD) soybean oil was essentially free of FFA (AV 0.03 mg KOH/g), which was expected as a result of the additional purification conducted on this commercially available commodity oil. The FFA content may be important in cases where chemical modification is to be conducted in the presence of catalysts that are sensitive to carboxylic acids. For instance, in the case of homogenous base-catalyzed methanolysis to yield fatty acid methyl esters, feedstocks with FFA contents greater than 0.5 wt% may result in reduced product yields (Naik et al., 2008). Consequently, acid-catalyzed pre-treatment to lower the FFA content of crude cress oil may be necessary prior to applications where certain acid-sensitive catalysts are to be employed (Rashid et al., 2008).

3.2. Tocopherol content

Cress oil contained high amounts of γ- (1422 ppm, Table 3) and δ- (356 ppm) tocopherols (see Fig. 1), but minimal quantities of α- (21 ppm), and β- (0 ppm) tocopherols. The unusually high percentage of γ-tocopherol may make cress oil a potentially useful industrial source of this natural antioxidant. For comparison, existing crude commodity vegetable oils with relatively high γ-tocopherol contents include corn (942 ppm), soybean (870 ppm), cottonseed (387 ppm), and palm (316 ppm) oils (Gunstone, 2004; Frankel, 2005). In the current study, RBD soybean oil contained only 537 ppm γ-tocopherol and 757 ppm combined tocopherols (Table 3). The total tocopherol content of cress oil was 1799 ppm,
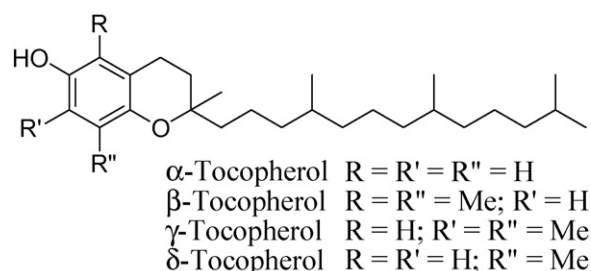

Fig. 1. Chemical structures of α-, β-, γ-, and δ-tocopherols found in cress, field pennycress, and soybean oils.

which was higher than that typically reported for crude soybean oil (1300–1600 ppm) (Jung et al., 1989). Other crude commodity vegetable oils normally contain less than 1000 ppm combined tocopherols, with the exception of corn (1175 ppm) and cottonseed (1000 ppm) oils (Gunstone, 2004).

Field pennycress oil primarily contained α- (714 ppm) and γ- (126 ppm) tocopherols, with β- (6 ppm) and δ- (5 ppm) tocopherols present in essentially trace amounts (Table 3). The total tocopherol content of field pennycress oil was 851 ppm, which was lower than cress oil but higher than other common crude commodity vegetable oils such as palm (642 ppm, combined), sunflower (546 ppm), and safflower (413 ppm) oils (Frankel, 2005).

3.3. Phytosterol content

The primary phytosterols detected in cress oil included sitosterol (5.82 mg/g), campesterol (3.95 mg/g), and avenasterol (3.44 mg/g), with cholesterol (0.50 mg/g), stigmasterol (0.30 mg/g), dihydrolanosterol (0.25 mg/g), and β-amyrin (0.16 mg/g) comprising the remaining steroidal constituents (Table 3). As is the case with most vegetable oils, sitosterol (see Fig. 2) was the major phytosterol found in cress oil. The combined phytosterol content of cress oil was 14.41 mg/g, which was in excess of the typical range of 1–5 mg/g for most vegetable oils (Gunstone, 2004). For comparison, crude commodity vegetable oils that are known to have high levels of phytosterols include corn (8–22 mg/g) and rapeseed (5–11 mg/g) oils (Gunstone, 2004).

Table 3
Tocopherol (ppm) and phytosterol (mg/g) contents of cress, field pennycress, and soybean oils.

Purity:Oil	Crude cress	Crude pennycress	RBD soybean
Tocopherols			
α-Tocopherol	21 (1)[a]	714 (7)	62 (2)
β-Tocopherol	0	6 (0)	11 (1)
γ-Tocopherol	1422 (11)	126 (3)	537 (5)
δ-Tocopherol	356 (4)	5 (0)	147 (3)
Σtoco[b]	1799 (9)	851 (6)	757 (5)
Phytosterols			
Cholesterol	0.50 (0.02)[b]	0.27 (0.01)	0
Brassicasterol	0	0.76 (0.01)	0
Campesterol	3.95 (0.22)	3.00 (0.07)	0.94 (0.04)
Stigmasterol	0.30 (0.02)	0.21 (0.01)	0.69 (0.02)
Sitosterol	5.82 (0.37)	3.88 (0.07)	2.09 (0.07)
Sitostanol	0	0	0.06 (0.01)
Avenasterol	3.44 (0.22)	0.44 (0.02)	0.15 (0.01)
Cycloartenol	0	0	0.24 (0.01)
Dihydrolanosterol	0.25 (0.02)	0	0
Citrostadienol	0	0	0.13 (0.01)
β-Amyrin	0.16 (0.01)	0	0
Σphyto[c]	14.41 (0.87)	8.55 (0.17)	4.29 (0.11)

[a] Values in parentheses are the standard deviation from the reported mean.

[b] Σtoco = sum of all tocopherols.

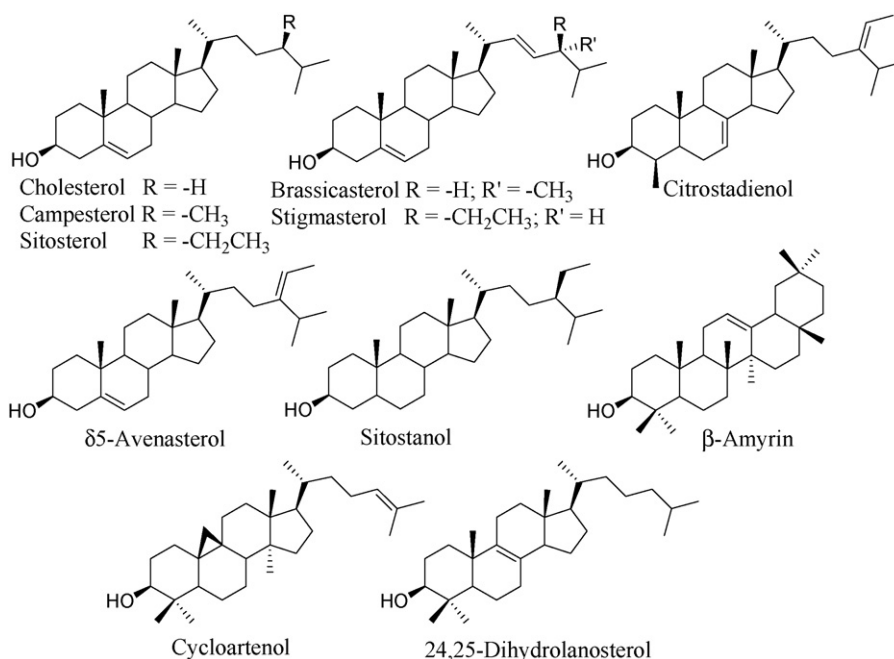[c] Σphyto = sum of all phytosterols.

Fig. 2. Chemical structures of phytosterols found in cress, field pennycress, and soybean oils.

The primary phytosterols detected in field pennycress oil were sitosterol (3.88 mg/g) and campesterol (3.00 mg/g), with brassicasterol (0.76 mg/g), avenasterol (0.44 mg/g), cholesterol (0.27 mg/g), and stigmasterol (0.21 mg/g) accounting for the remaining phytosterol content (Table 3). The combined phytosterol concentration of crude field pennycress oil (8.55 mg/g) was less than that found for crude cress oil, but still higher than RBD soybean oil (4.29 mg/g, Table 3). Field pennycress and cress oils were both high in sitosterol and campesterol and contained nearly the same phytosterols, with the exception of brassicasterol in the case of field pennycress oil and dihydrolanosterol and β-amyrin in the case of cress oil. It is interesting that cress oil had no detectable levels of brassicasterol, which typically comprises 5–20% of phytosterols in oils from other plants in the Brassicaceae family, such as rapeseed, canola, and mustard oils (Kochhar, 1983; Phillips et al., 2002). Bettach et al. (1997) also did not detect brassicasterol in the unsaponifiable fraction of cress oil or in *Diplotaxis tenuisiliqua*, another member of the Brassicaceae family.

3.4. Physical properties

Field pennycress oil exhibited the most desirable low temperature fluidity, as indicated by CP and PP values of −25 and −28 °C versus −7 and −9 °C for soybean oil and 3 and −11 °C for cress oil. Based on these results, it is expected that field pennycress oil would be better suited for industrial applications where sub-ambient conditions are prevalent. The relatively low level of saturated FA contained in field pennycress oil (4.6 wt%; Table 1) in comparison to cress (15.2 wt%) and soybean (15.9 wt%) oils is attributed to its enhanced low temperature operability. A previous study elucidated a statistically significant relationship between saturated FA content and low temperature properties (Moser, 2008a). In the case of fatty acid methyl esters, compounds of similar chain length but increasing levels of unsaturation display lower melting points (mp), as evidenced by the mp of methyl esters of stearic (mp 39 °C), oleic (mp −20 °C), linoleic (mp −35 °C), and linolenic (mp −52 °C) acids. The propensity of chemical species with greater amounts of polyunsaturation to exhibit lower mp also explains

why soybean oil displayed superior low temperature properties in comparison to cress oil: the polyunsaturated FA content of soybean oil (59.8 wt%) was significantly higher than that for cress oil (37.7 wt%).

Cress oil displayed greater stability to oxidation than field pennycress oil, as indicated by IP values of 12.3 and 5.0 h (Table 2), respectively, according to official method EN 14112 (CEN, 2003). The rate of autoxidation is dependant on the number and location of methylene-interrupted double bonds contained within fatty materials and by the presence of antioxidants. Polyunsaturated materials are particularly vulnerable to autoxidation, as evidenced by the relative rates of oxidation of unsaturated esters: 1 for ethyl oleate, 41 for ethyl linoleate, and 98 for ethyl linolenate (Holman and Elmer, 1947). Although cress oil contained a higher amount of linolenic acid (29.3 wt%) than field pennycress oil (11.8 wt%), the higher native content in cress oil of oxidation inhibitors such tocopherols (Evans et al., 2002; Kamal-Eldin, 2006; Moser, 2008b) imparted greater resistance to oxidation. Differences in the tocopherol composition may also explain the difference in stability between cress and field pennycress oils. The major tocopherol constituent in cress oil was γ-tocopherol, which in bulk oils is a better antioxidant than α-tocopherol, the main tocopherol homologue in field pennycress oil (Huang, 1995; Lampi et al., 1999). The oxidative stability of RBD soybean oil (8.3 h) was between that of cress and field pennycress oils.

Field pennycress oil had an IV of 118 (Table 2), which was lower than that of cress (130) and soybean (130) oils. The IV, which is an indicator of the level of unsaturation, is not a good predictor of oxidative stability of vegetable oils. For instance, field pennycress oil had the lowest IV (lowest double bond content), but exhibited the worst oxidative stability of the oils examined in this study. Additionally, cress and soybean oils had identical IV, but significantly different oxidative stabilities. These observations are supported by a previous study that questions the utility of IV as a predictor of oxidative stability (Knothe, 2002). Furthermore, IV cannot adequately account for the presence of minor constituents such as antioxidants that may strongly influence stability to oxidation in vegetable oils.

The kinematic viscosity of field pennycress oil was higher than that for cress and soybean oils over a wide range of temperatures (25–100 °C; Table 2) as a result of its relatively high erucic acid content. Kinematic viscosity is dependent upon the constituent FA contained within vegetable oils. Fatty acids of longer chain length are progressively more viscous, as evidenced by the kinematic viscosities (40 °C) of methyl esters of palmitic (3.67 mm^2/s), oleic (4.51 mm^2/s), gondoic (5.77 mm^2/s), and erucic (7.33 mm^2/s) acids (Knothe and Steidley, 2005). The kinematic viscosity of field pennycress oil at 25, 40, and 100 °C was 70.01, 40.97, and 9.39 mm^2/s (Table 2), respectively, which resulted in a calculated VI of 224. Cress oil exhibited lower kinematic viscosities at 25, 40, and 100 °C (44.12, 28.35, and 7.35 mm^2/s, respectively) and provided a calculated VI of 244. Soybean oil displayed kinematic viscosities and VI intermediate to those of cress and field pennycress oils. All oils yielded high VI, which is desirable in industrial applications where substantial changes in viscosity over a broad range of temperatures are undesirable. Higher VI oils can be used to formulate multi-purpose lubricants. The specific gravities of cress, field pennycress, and soybean oils were nearly indistinguishable at both 25 and 40 °C (see Table 2), with soybean oil yielding only slightly higher values at both temperatures. These results demonstrate that the nature of the FA constituents, at least in this case, do not significantly impact density and, by extension, specific gravity of vegetable oils.

An advantage to vegetable oils over that of refined petroleum products is superior lubrication properties (lubricity) resulting from structural features such as unsaturation and the presence of heteroatoms not found in hydrotreated petroleum-derived materials. For example, in a recent study, vegetable oils, biodiesel, and vegetable oil derivatives exhibited superior lubricity in comparison to unadditized conventional petroleum-derived ultra low sulfur diesel fuel (ULSD, <15 ppm S) (Moser et al., 2008). In the present study, all vegetable oils displayed similar lubricities (124–133 μm by HFRR; Table 2), which were significantly better (shorter wear scar lengths) than the wear scar generated according to ASTM D6079 by ULSD (551 μm; Moser et al., 2008) in a prior investigation.

4. Conclusions

Crude field pennycress oil exhibited excellent low temperature fluidity, whereas crude cress oil was more stable to oxidation and over a wide range of temperatures displayed lower kinematic viscosities. Both oils afforded high viscosity indices and excellent lubricities. The acid and iodine values of crude field pennycress oil were lower than those for crude cress oil. These oils compare favorably, with the exception of kinematic viscosity in the case of field pennycress oil, to soybean oil with respect to these properties.

The primary fatty acids in cress oil were oleic (30.6 wt%) and linolenic acids (29.3 wt%), whereas field pennycress oil was principally composed of erucic (32.8 wt%) and linoleic (22.4 wt%) acids. Cress oil contained high concentrations of γ- (1422 ppm) and δ- (356 ppm) tocopherols, whereas α-tocopherol (714 ppm) was the primary homologue in field pennycress oil. The overall tocopherol concentrations of cress and field pennycress oils were 1799 and 851 ppm, respectively. The primary phytosterols in both oils were sitosterol and campesterol, with avenasterol also present in significant quantity in cress oil. The overall phytosterol concentration in cress oil (14.41 mg/g) was greater than that for field pennycress (8.55 mg/g) oil.

References

American Oil Chemists Society (AOCS), 1999a. Determination of tocopherols and tocotrienols in vegetable oils and fats by HPLC. In: Official Methods and Recommended Practices of the American Oil Chemists Society, 5th ed. AOCS, Champaign, IL, p. Ce 8-89.

American Oil Chemists Society (AOCS), 1999b. Specific gravity of oils and liquid fats. In: Official Methods and Recommended Practices of the American Oil Chemists Society, 5th ed. AOCS, Champaign, IL, p. Cc 10a-25.

American Oil Chemists Society (AOCS), 1999c. Acid value. In: Official Methods and Recommended Practices of the American Oil Chemists Society, 5th ed. AOCS, Champaign, IL, p. Cd 3d-63.

American Oil Chemists Society (AOCS), 1999d. Calculated iodine value. In: Official Methods and Recommended Practices of the American Oil Chemists Society, 5th ed. AOCS, Champaign, IL, p. Cd 1c-85.

American Society for Testing Materials (ASTM), 2001. Standard test method for pour point of petroleum products (automatic pressure pulsing method) ASTM (D5949-01). In: ASTM Annual Book of Standards. ASTM International, West Conshohocken, PA.

American Society for Testing Materials (ASTM), 2004a. Standard Practice for calculating viscosity index from kinematic viscosity at 40 and 100 °C. In: ASTM Annual Book of Standards. ASTM International, West Conshohocken, PA.

American Society for Testing Materials (ASTM), 2004b. Standard test method for evaluating lubricity of diesel fuels by high frequency reciprocating rig (HFRR) ASTM (D6079-04). In: ASTM Annual Book of Standards. ASTM International, West Conshohocken, PA.

American Society for Testing Materials (ASTM), 2006. Standard test method for kinematic viscosity of transparent and opaque liquids (and calculation of dynamic viscosity) ASTM (D445-06). In: ASTM Annual Book of Standards. ASTM International, West Conshohocken, PA.

American Society for Testing Materials (ASTM), 2007. Standard test method for cloud point of petroleum products (constant cooling rate method) ASTM (D5773-07). In: ASTM Annual Book of Standards. ASTM International, West Conshohocken, PA.

Bettach, N., Banoub, J., Delmas, M., 1997. Comparative study of the seeds of the Lepidium, Eruca, Diplotaxis and Sinapis Crucifers II. Description of the Sterols. Oléagineux, Corps Gras. Lipides 4 (2), 152–156.

Chung, J., Babka, H.L., Graef, G.L., Staswick, P.E., Lee, D.J., Cregan, P.B., Shoemaker, R.C., Specht, J.E., 2003. The seed protein, oil, and yield QTL on soybean linkage group I. Crop Sci. 43, 1053–1067.

Dolya, V.S., Koreshchuk, K.E., Shkurupii, E.N., Kaminskii, N.A., 1976. Oils of three representatives of the family Cruciferae II. Chem. Nat. Comp. 10, 447–449.

Dornbos Jr., D.L., Mullen, R.E., 1992. Soybean seed protein and oil contents and fatty acid composition adjustments by drought and temperature. J. Am. Oil Chem. Soc. 69, 228–231.

Dutta, P.C., Normén, L., 1998. Capillary column gas-liquid chromatographic separation of Δ5-unsaturated and saturated phytosterols. J. Chromatgr. A 816, 177–184.

European Committee for Standardization (CEN), 2003. Fat and oil derivatives. In: Fatty Acid Methyl Esters (FAME). Determination of Oxidative Stability (Accelerated Oxidation Test) EN (14112:2003). CEN, Brussels, Belgium.

Evangelista, R.L., 2009. Oil extraction from lesquerella seeds by dry extrusion and expelling. Ind. Crops Prod. 29, 189–196.

Evans, J.C., Kodali, D.R., Addis, P.B., 2002. Optimal tocopherol concentrations to inhibit soybean oil oxidation. J. Am. Oil Chem. Soc. 79, 47–51.

Frankel, E.N., 2005. Lipid Oxidation. The Oily Press, Bridgewater, p. 225.

Gokavi, S.S., Malleshi, N.G., Guo, M., 2004. Chemical composition of garden cress (Lepidium sativum) seeds and its fractions and use of bran as a functional ingredient. Plant Food. Hum. Nutr. 59, 105–111.

Gunstone, F.D., 2004. The Chemistry of Oils and Fats. Sources, Composition, Properties and Uses. CRC Press, Boca Raton.

Holman, R.A., Elmer, O.C., 1947. The Rates of Oxidation of Unsaturated Fatty Acids and Esters. J. Am. Oil Chem. Soc. 24, 127–129.

Huang, S.-W., 1995. Effects of individual tocopherols and tocopherol mixtures on the oxidative stability of corn oil triglycerides. J. Agric. Food Chem. 43, 2345–2350.

Jung, M.Y., Yoon, S.H., Min, D.B., 1989. Effects of processing steps on the contents of minor compounds and oxidation of soybean oil. J. Am. Oil Chem. Soc. 66, 118–120.

Kamal-Eldin, A., 2006. Effect of fatty acids and tocopherols on the oxidative stability of vegetable oils. Eur. J. Lipid Sci. Technol. 58, 1051–1061.

Knothe, G., 2002. Structure indices in fatty acid chemistry how relevant is the iodine value? J. Am. Oil Chem. Soc. 79, 847–854.

Knothe, G., Steidley, K.R., 2005. Kinematic viscosity of biodiesel fuel components and related compounds influence of compound structure and comparison to petrodiesel fuel standards. Fuel 84, 1059–1065.

Lampi, A.-M., Kataja, L., Kamal-Eldin, A., Piironen, V., 1999. Antioxidant activities of α- and γ-tocopherols in the oxidation of rapeseed oil triacylglycerols. J. Am. Oil Chem. Soc. 76, 749–755.

Moser, B.R., 2008a. Influence of blending canola, palm, soybean, and sunflower oil methyl esters on fuel properties of biodiesel. Energy Fuel 22, 4301–4306.

Moser, B.R., 2008b. Efficacy of myricetin as an antioxidant in methyl esters of soybean oil. Eur. J. Lipid Sci. Technol. 110, 1167–1174.

Moser, B.R., Cermak, S.C., Isbell, T.A., 2008. Evaluation of castor and lesquerella oil derivatives as additives in biodiesel and ultra low sulfur diesel fuel. Energy Fuel 22, 1349–1352.

Naik, M., Meher, L.C., Naik, S.N., Das, L.M., 2008. Production of biodiesel from high free fatty acid Karanja (*Pongamia pinnata*) oil. Biomass Bioenerg. 32, 354–357.

Phillips, K.M., Ruggio, D.M., Toivo, J.I., Swank, M.A., Simpkins, A.H., 2002. Free and esterified sterol composition of edible oils and fats. J. Food Comp. Anal. 15, 123–142.

Rashid, U., Anwar, F., Moser, B.R., Knothe, G., 2008. *Moringa oleifera* oil: a possible source of biodiesel. Bioresour. Technol. 99, 8175–8179.

Vaughn, S.F., Isbell, T.A., Weisleder, D., Berhow, M.A., 2005. Biofumigant compounds released by field pennycress (*Thlaspi arvense*) seedmeal. J. Chem. Ecol. 31, 167–177.